Introduction

Bivalve molluscs (oysters, mussels, clams and scallops) form a significant part of the world's fisheries production. In 2000 landings of bivalves from capture fisheries and aquaculture operations totalled 14 204 152 tonnes (Figure 1). During the decade from 1991 to 2000 there was a continuing increase in production of bivalves, and landings more than doubled from 6.3 million tonnes in 1991 to 14 million tonnes in 2000.

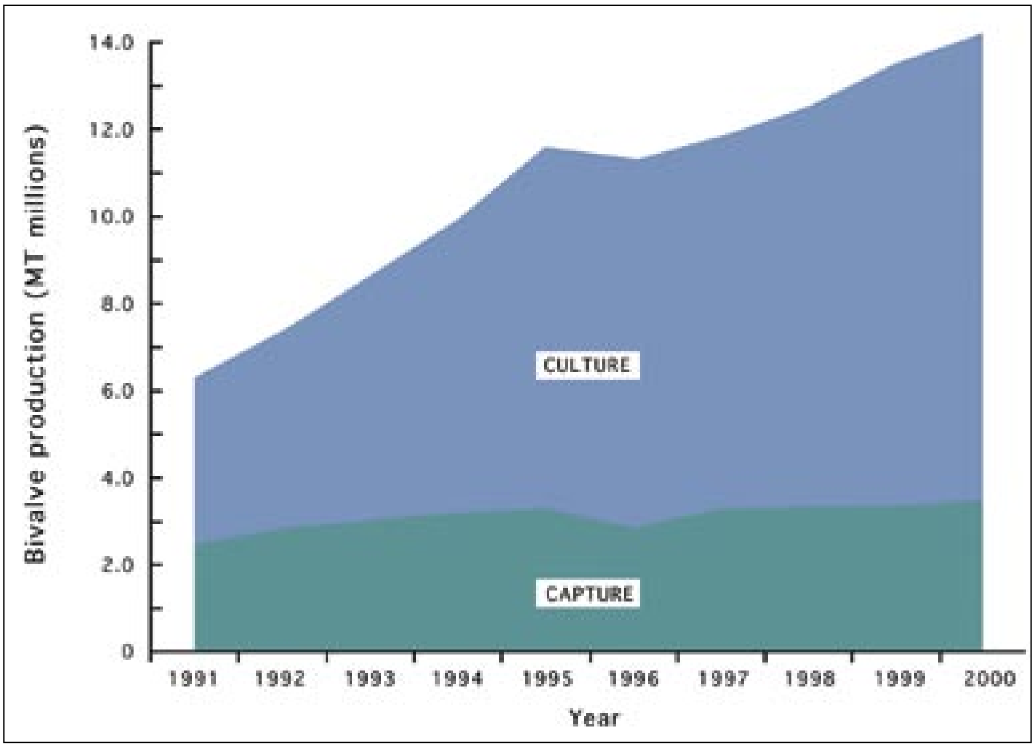

Figure 1: Production (in millions of tonnes) of bivalves from capture fisheries and aquaculture during the decade from 1991 to 2000 (from FAO Yearbooks of Fishery Statistics).

The global trend in the growth of human consumption of seafood will undoubtedly continue. Seafood constitutes an important and essential part of the diet of many people in the world and the need for increased production in these countries will persist as the human population expands. In some countries, seafood is recognized as an important and healthy part of the human diet and demand for seafood products in these countries will also grow. Most of the demand for seafood is and will continue to be for finfish, but the production and harvest of molluscs, particularly bivalves, will also be important in meeting rising demand. While the harvest of natural bivalve stocks will continue to prove significant, many wild stocks are probably already being harvested at or near maximum sustainable limits, in some places and stocks may even be overharvested. Aquaculture is the alternative to the harvest of wild stocks.

During the period 1991–2000, bivalve landings from capture fisheries increased only slightly from about 2.5–3.5 million tonnes, while landings from culture operations more than doubled during this same period, increasing from 6.3–14 million tonnes (Figure 2). About 75% of the world's bivalve production in 2000 resulted from some form of culture operation.

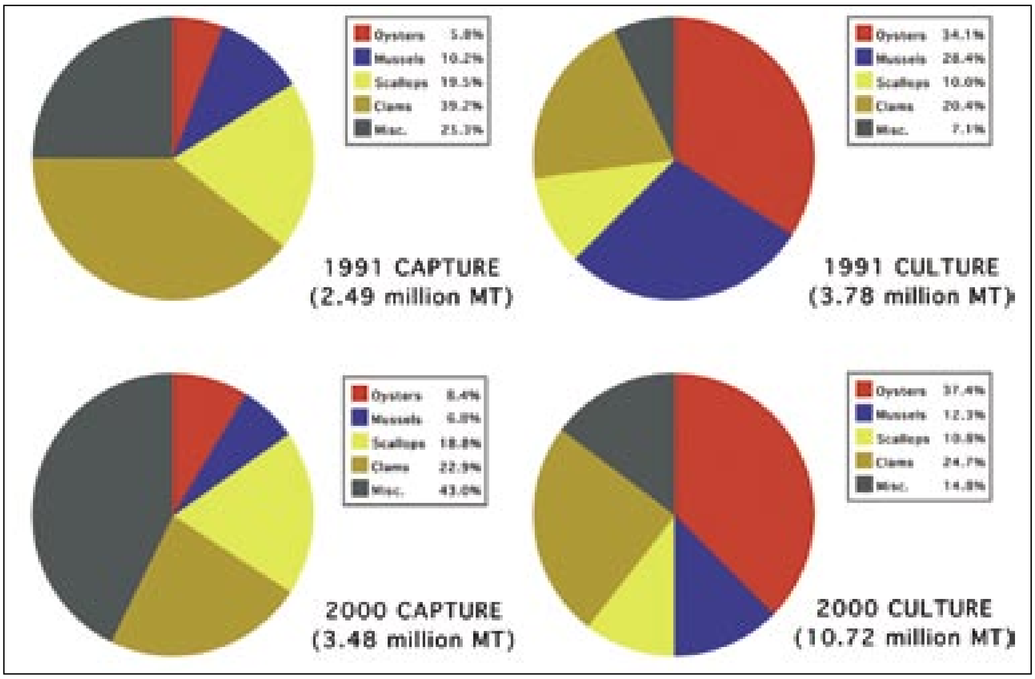

Figure 2: A comparison of production from capture fisheries and aquaculture broken down into the relative contributions of the major groups of bivalves in 1991 and 2000.

Bivalves are ideal animals for aquaculture: they are herbivores requiring no additional feeding apart from the natural algae content of seawater and generally minimum husbandry. Altough they have been cultured for hundreds of years advances in culture technology in recent years have led to significantly higher production. Continued improvements in culture methodology and technology will be required to meet increasing demand and also to make bivalve culture more economically attractive to both investors and people who wish to become shellfish farmers. The need for greater efficiency in culture operations will become even more essential in the future as areas where shellfish farming can be undertaken in the world are limited and may become more restricted as human populations and coastal urbanization grow.

A primary requisite in any culture or farming operation is an abundant, reliable and inexpensive supply of juveniles (seed). At present, most bivalve culture operations in the world obtain their seed by collecting natural sets. A substrate (cultch) is placed in breeding areas to collect metamorphosing larvae and the juveniles collected are transferred to growing areas for culture (growout) to market size. In other operations, juveniles are gathered from areas of natural abundance and are transported to growing areas that may be distant from the source of the seed. Collection of juveniles from areas of natural recruitment will continue to be important in bivalve culture operations worldwide and undoubtedly can be expanded in some areas to meet increased seed requirements for culture operations. The importance of these breeding areas must be recognized and every effort made to conserve them.

In many other culture locations, natural breeding areas do not exist to provide seed or, if they exist, they cannot produce sufficient seed to meet growout requirements, or breeding is erratic and an annually reliable source of seed cannot be guaranteed. There are other considerations that may make collection of natural sets unsuitable as the supply for culture operations. Growers in some areas may wish to develop and culture a particular race or strain of bivalve to suit their individual needs and juveniles for such operations are not available from the local or nearby environment. Another consideration is that growers may wish to introduce a non-indigenous (exotic) species but a seed source is not available. The alternative to collection of natural sets of bivalves is to produce seed in a hatchery. Bivalve hatcheries have existed for over half a century and are well-established in several countries. They are an integral part of many culture operations and the major or sole source of seed. In the future, bivalve hatcheries will undoubtedly become more important to culture operations as shellfish farming becomes more specialized and the need for seed increases.

Hatcheries have several advantages over collection of natural sets. They are reliable and can supply growers with their seed requirements when it is convenient to them – often much earlier in a growing season than natural sets would occur. They can supply seed that is not available to farmers from natural sets, for example, genetic strains that have improved biological characteristics for farming operations in local areas or a supply of exotic bivalve seed. Cost is the main disadvantage of producing seed in hatcheries: it is more expensive to propagate seed in a facility than collecting it from natural sets. Although economic factors have probably caused the demise of some bivalve hatcheries in the past, recent improvements in technology have greatly enhanced both reliability and financial viability whereby it is possible to produce seed at competitive prices. In some parts of the world hatcheries are the only source of seed for the commercial culture industry but there is ample scope to make them more efficient and more widely accepted as the preferred source of seed.

Constructing and operating a bivalve hatchery is a large and expensive undertaking and careful thought must be given to the developmental phase of a new venture otherwise it will likely end in failure. There is no single plan to construct and operate a bivalve hatchery; indeed many have begun as a small operation and simply grown as markets for their products expanded. Hatcheries vary greatly in their design, configuration and construction from site to site depending on species cultured, target production levels and, most of all, the local conditions and personal preferences of owners/operators. However, the basics for any bivalve hatchery are the same and include a method to condition and spawn adults, rear and set larvae, rear the juveniles to an acceptable size, together with a facility to produce large quantities of food (algae) to feed all stages of the production cycle. Although these essential elements are the same in every hatchery, there are variations in technology and the efficiency of each phase of operation must continually be improved to make hatcheries increasingly more profitable.

This publication is intended to serve as a "how to" manual for bivalve hatcheries. While other documents also describe bivalve hatcheries many are becoming out-of-date and do not include the most recent technological improvements. This manual is intended as a practical introduction to the fundamentals of hatchery operation for new entrants in this field. It will also allow a potential investor to assess whether he/she wishes to build and operate a bivalve hatchery and become involved in the business of producing seed for the culture industry. This is not meant to be a scientific publication in the accepted sense and much of the content is based on the authors' own experience gained over a collective period of 80 or more years. Although there is an extensive literature pertinent to bivalve hatcheries, many of the more practical publications have had limited circulation and are out of print or are only available through specialist library services. Many readers may not be able to access such written material and so efforts have been made to make this manual as comprehensive and available as possible. Rather than including extensive references in the text, a suggested reading list is given at the end of each major section to provide further sources of information on particular subjects and aspects of operation.

Part 1

Site selection, hatchery design and economic considerations

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1.1 SITE SELECTION

1.1.1 Introduction

Selecting the proper site for a bivalve hatchery is the most important consideration when deciding to build a hatchery, yet it is a factor that has often been overlooked when some hatcheries were built. Several factors may have contributed to the inappropriate location of a facility including the lack of one or more of the components of the essential infrastructure, e.g. land availability at reasonable cost, the local availability of electricity and freshwater, a qualified labour force, or good communications. A further consideration has often been that an individual or company may have wished to build a hatchery at a site adjacent to an existing bivalve growout operation. In such cases the hatchery became an add-on feature to an existing operation. Yet, another factor is that an individual or company may own or have rights to a particular location and it proves to be the only place where a hatchery could conveniently be built. While it is true that it may be impossible to build a hatchery at an ideal location, nevertheless certain criteria must be met or a hatchery will likely be doomed to failure.

1.1.2 Considerations

1.1.2.1 Government regulations

The first consideration is to determine if government regulations permit construction of a bivalve hatchery at the desired site. This can be done quickly by making enquiries of local, state, provincial or federal authorities. If regulations do not permit construction of a hatchery at the desired site one must decide if it is preferable to find another location where construction is permitted, or attempt to change existing government regulations to allow construction at the desired site.

It is likely that a number of permits and licences will be required to ensure compliance with local building codes and national and local environmental regulations before any construction is allowed. This can be a lengthy, costly and time consuming process and may require an assessment of the potential impact of the hatchery on the local environment before permission is either granted or not granted to begin the construction phase.

1.1.2.2 Seawater quality

Before committing to what is considered to be a suitable location for a hatchery it is of paramount importance to ensure that good quality seawater exists year-round at the prospective site. This point cannot be overemphasized. If a good seawater source is not available, it will be difficult, if not impossible, to develop an efficient and profitable hatchery operation. For this reason every effort should be made to obtain as much information as possible about the quality of the seawater throughout the year at a potential site - or sites. Information is required not only for surface waters but also for the entire water column, since thermoclines may develop or upwelling may occur periodically. If previous oceanographic surveys have been undertaken in the area, copies of the data should be examined. If such surveys have not been undertaken, one should be prepared to undertake a detailed sampling of the waters at the proposed site for at least a year.

Environmental parameters of seawater that need to be examined will depend in part on geographic location and the intended species for culture. Bivalve larvae as well as juveniles and adults have strict physiological requirements, such as water temperature, salinity and oxygen levels and these must be maintained in a hatchery operation. Water temperatures are higher in the tropics than in temperate regions and indigenous bivalves are well adapted to tolerate these conditions. But in a hatchery situation temperatures must not be allowed to drop too low or larval and juvenile survival and growth will be adversely affected. In temperate areas water temperatures must not be allowed to exceed upper or lower lethal levels to larvae and juveniles. Salinity can vary widely and tolerance to these fluctuations differs among bivalve species. Some require high oceanic levels of salinity while euryhaline (estuarine and brackish water) species exhibit much wider tolerance. Periods of heavy rainfall may not only cause periods of low salinity, but heavy associated runoff can increase quantities of silt and other materials which may lead to problems in a hatchery. Dense concentrations (blooms) of some marine algal and bacteria species may release toxic substances that may cause reductions in both the survival and growth of bivalve larvae or juveniles, or mass mortalities in extreme cases. As much data as possible on these parameters should be collected prior to deciding on the adequacy of a site for a bivalve hatchery. Remedial measures to improve inadequate quality seawater can be extremely costly and may adversely effect the profitability of a venture.

Locations possibly influenced by effluents discharged from industrial plants should be avoided. The lethal and sublethal effects of many industrial pollutants are not completely understood, nor are the additive effects they may exert when several industries are discharging a range of potentially toxic wastes in nearby waters. Effects of such effluents can be extremely damaging to bivalve larvae. For example, an anti-fouling ingredient added to marine paints, tributyltin (TBT), has been found to be highly lethal to bivalve larvae even at concentrations of a few parts per billion. Drawing a seawater supply from the vicinity of marinas and commercial docks needs to be avoided. If feasible it is advisable to undertake bioassay studies using bivalve embryos to help determine the quality of the water at the potential hatchery site. The presence of deleterious materials may be transitory or seasonal in nature, so sampling for bioassays should be carried out over a period of at least a year and be done preferably on a weekly basis.

Agricultural – forestry included – and domestic sources of pollution should also be avoided. It has recently been shown that runoff from some cultivated lands can carry concentrations of pesticides at levels deleterious to the growth and survival of bivalve larvae. Domestic pollution may not only contain pollutants that are toxic to bivalve larvae but the high organic content can cause depletion of oxygen levels and increased levels of bacteria that could also lead to reduced growth and mortalities of larvae.

Another consideration when deciding upon the location of a bivalve hatchery is whether "civilization" will soon encroach on the site. Urbanization with its ancillary problems is one of the main concerns in bivalve culture. If the site will soon be encompassed by urbanization then every effort must be made to ensure that sources of potential pollution will be kept to a minimum. This will require working closely with planners and developers.

1.1.2.3 Siting the hatchery

The hatchery should be located close to the ocean so that the distance required to pump water is kept to a minimum. This negates the necessity of having to maintain great lengths of pipe. It should also be located as close to sea level as possible to avoid problems of pumping water any great vertical distance. If fluctuations in surface seawater temperature and salinity occur regularly, the intakes for the pipes will need to be located at depth (up to 20 m below the surface) to maintain more constant water temperature and salinity. Depending on the nature of the geological strata, it may be possible to drill wells close to the shore to access seawater aquifers. A water source of this nature will be at a more constant temperature year-round and will already be pre-filtered by percolation through the strata. It may, however, require oxygenating before use. It is always wise to consult with a suitably qualified engineer when making decisions on the best methodology and technology to procure the water supply.

Sufficient area needs to be available at the site to accommodate the hatchery and ancillary buildings and also to allow for any future expansion. The need for adequate surveillance should also be considered.

Other considerations that need to be kept in mind for a site include an adequate supply of electrical power, a source of freshwater and a skilled labour force to operate the hatchery. Good communications should exist so that required materials and supplies can be acquired quickly and larvae and seed can be quickly shipped to their various destinations. The proximity of institutions such as universities, government laboratories and libraries should also be considered since such resources can be of great assistance in operations and in helping towards solutions to problems that may arise.

It is a worthwhile preliminary to prepare a check list of parameters that must be met, or at least reviewed, when considering a site for a bivalve hatchery and work through the list to ensure the site meets as many of the requirements as possible.

1.2 HATCHERY DESIGN CONSIDERATIONS

1.2.1 Introduction

There is no rigid design for a bivalve hatchery. The layout of hatcheries varies from site to site, with species produced, geographic location, funds available, the target production species and personal preferences (Figure 3). Some hatcheries are small and supply seed for their own bivalve on-growing culture operations. Others are large and may only produce seed for sale, or they may produce seed for their own operations and also an excess to sell to other growers. Hatcheries may or may not include a nursery component and some may only produce mature larvae for shipment elsewhere while others may grow and supply seed varying in size from 1 to 12 mm shell length. Much depends upon the nature, requirements and the level of sophistication of the growout operations that collectively make the customer base.

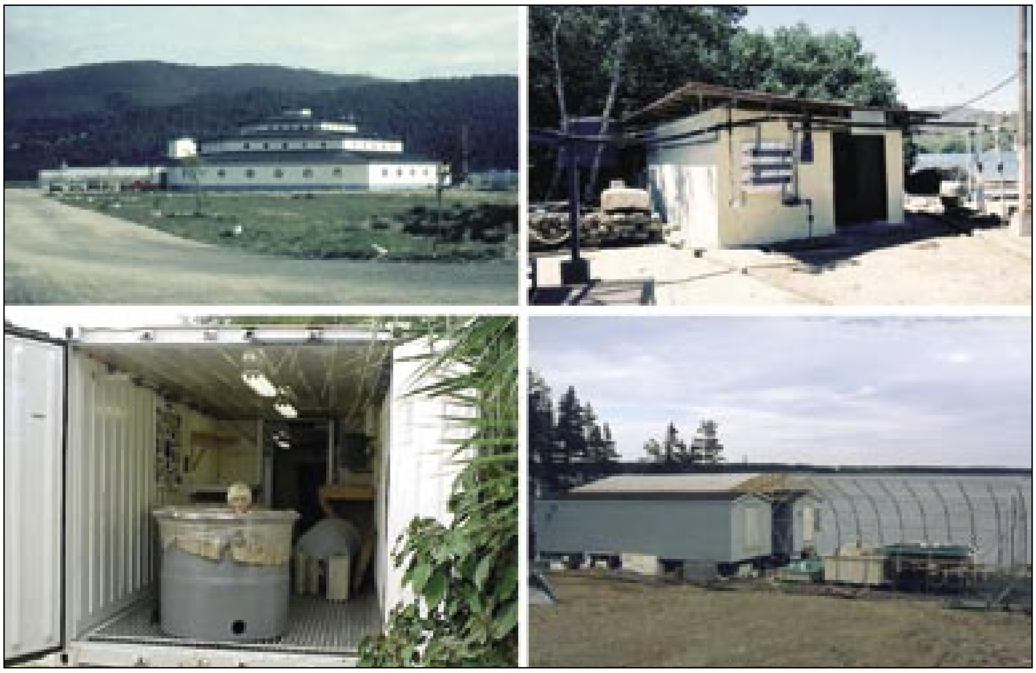

Figure 3: A selection of photographs of hatcheries depicting the variability in size and sophistication of construction that exists around the world. Clockwise from top left: Tinamenor S.A. (Pesues, Spain), Turpiolito hatchery, (Gulf of Cariaco, Venezuela), Bermuda Biological Station's scallop hatchery based on insulated cargo containers and the SMS oyster hatchery (Point Pleasant, Nova Scotia, Canada).

Many hatcheries were built with little advance planning or forethought for possible future development. A hatchery was built to produce a required quantity of seed and when the initial objective was achieved a decision was made to expand and add extra capacity. The resulting facility is often neither efficient nor worker friendly. Other hatcheries were built to produce seed of a single species but other species are produced now and the resulting hatchery is somewhat inefficient in its new role.

Considerable time will be saved and many frustrations avoided if a hatchery is carefully planned before construction begins. Several considerations must be remembered when designing a hatchery and two are of great importance. Firstly, the hatchery operation must be worker friendly and efficient to make the operation as profitable as possible, and secondly, the need for future expansion must be kept in mind.

There are two basic parts to a bivalve hatchery, the salt water system and the physical plant.

1.2.2 Seawater system

The need for a supply of high quality seawater was previously discussed. It is important to ensure that the seawater source and system to pump and treat it is located conveniently close to the hatchery and optimum use made of it to keep capital and operating costs to a minimum.

The hatchery should be located as close to sea level as possible to avoid lifting water. Intakes for the seawater should be as short as possible and conveniently located so they can be serviced and maintained with minimum effort. Intakes for the salt water should be located at depth to avoid fluctuations in temperature and salinity and also to reduce the number of organisms and amount of detritus that will enter the system. In temperate areas, intakes should be located below any thermocline that occurs in summer to reduce temperature variability. In areas where periods of heavy rain occur, the intakes should be deep enough to avoid sudden fluctuations in salinity and heavy siltation that may occur with the rains. Intakes at depth avoid major plankton blooms, some of which may be harmful to bivalve larvae and also greatly diminishes the number of fouling organisms entering the system. Fouling organisms can settle in pipes and greatly reduce water flow into the hatchery. Many of the above sources of variability can be avoided by accessing seawater from drilled wells. This possibility should be investigated before any other solution is considered.

Size of pumps and the diameter of the pipes required will depend on the scale of the operation and the volumes of seawater required to meet all aspects of production. Pumps are available through commercial outlets and the type and size of pump required can be determined after discussions with dealers. It is important to ensure that surfaces that come into contact with the seawater are non toxic. Most plastics, cast iron and certain grades of stainless steel are suitable. Pumps that contain mild steel or brass components should be avoided.

Seawater pumped directly from the ocean is first passed through sand filters that filter out most particulate material greater than 20-40 µm in size (Figure 4). A well maintained sand filter will remove the major portion of detritus and organisms from the water that may interfere with bivalve larvae. It also eliminates many of the fouling organisms that could settle and grow in pipes in the hatchery. They not only can cause problems with water flow but when they die they can produce anaerobic conditions that can be toxic to bivalve larvae. They may also harbour and eliminate bacteria that can be deleterious to larvae. Sand filters are commercially available and are the same or similar to those used to filter water in swimming pools. A series of two or more such filters are generally installed and they are regularly back-flushed to avoid clogging of the filter media. Other types of filters may be used depending on personal preference and cost considerations. Self-cleaning, rotating drum filters offer an alternative to remove larger particulate material and large surface area cartridge or bag filters are available and are extremely effective in removing smaller sized particulates.

Another method to obtain seawater for a hatchery is to pump it from seawater wells. This has become the preferred method for hatcheries to obtain their water supply in recent years. A well is dug or drilled close to the hatchery and is deep enough to provide a sufficient supply of seawater for the hatchery. Water from such wells is of high quality and generally has a constant temperature and salinity. It has already been filtered through sedimentary or porous rock, contains little detritus and few, if any, fouling organisms. Water abstracted in this way requires little if any further filtration. Constructing seawater wells can be expensive initially but the high capital cost is offset by reduced operating costs.

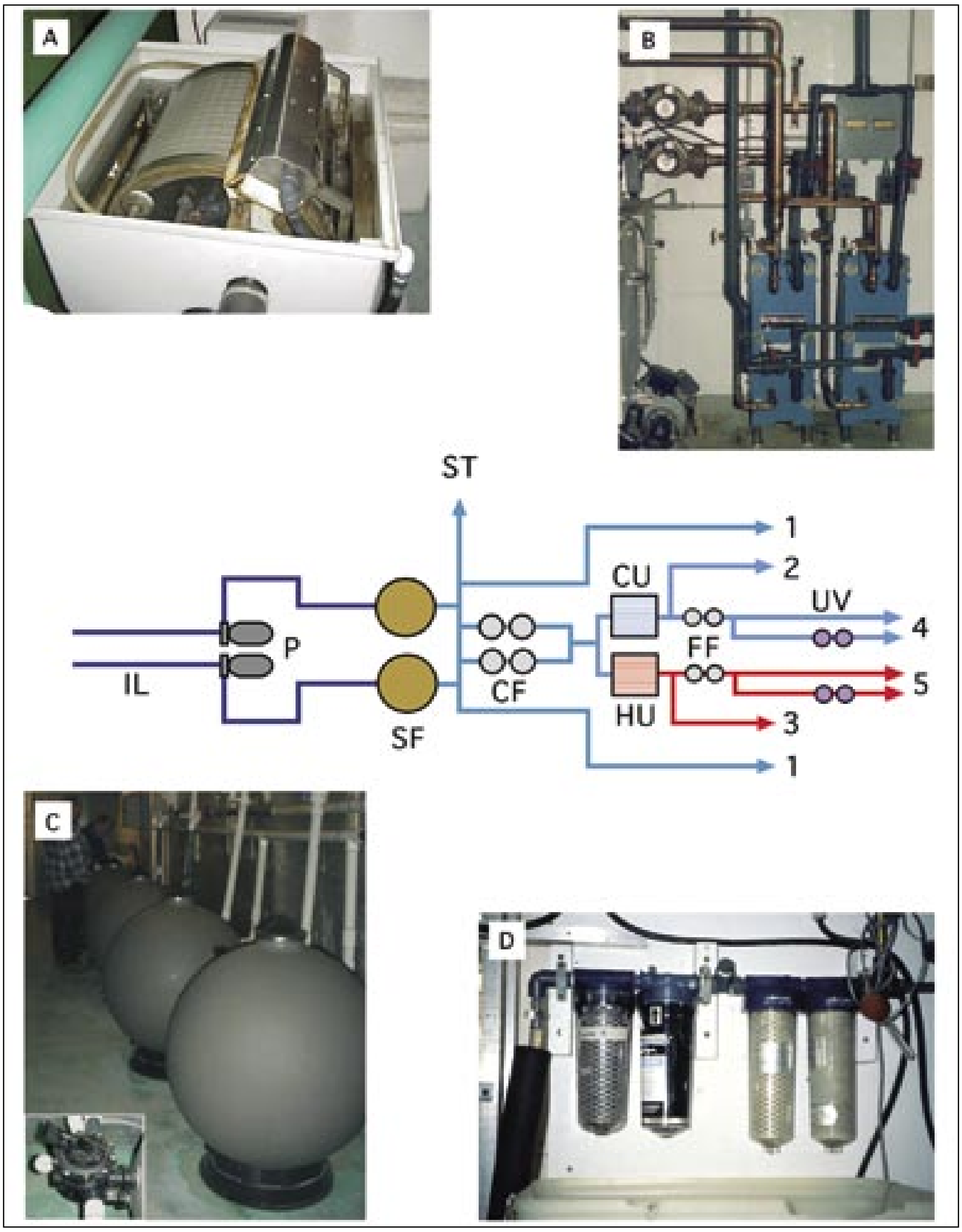

Figure 4: A diagram of the various stages of seawater treatment for hatchery usage from the intake pipes (IL) to the points at which water is used in the different aspects of the operation (1 to 5). Key: P – seawater pumps; SF – sand filters (photograph **C**) or alternatively self-cleaning drum filters (photograph **A**); ST – to storage tanks (if required); CF – cartridge filters of 20 µm and 10 µm; CU – seawater chilling unit (if required); HU – seawater heating unit (if required – photograph **B**); FF – final filtration (5 µm and 1 or 2 µm - photograph **D**); UV – ultra-violet light disinfecting units (if required).

A guide to typical usages (treatment levels vary from hatchery to hatchery):

1 – Unheated, sand-filtered water for broodstock and larger juveniles (if water requires to be heated, then 3).

2 – Chilled seawater filtered to 10 µm for spawning broodstock or for large-scale algal culture of hardy species. Chilled (or ambient temperature) seawater is often mixed with heated seawater to provide intermediate temperatures for a variety of purposes.

3 – Heated seawater filtered to 10 µm for conditioning and spawning broodstock and for growing larger spat. Some hatcheries have a separate heating system for either unfiltered or sand-filtered seawater for broodstock conditioning.

4 – Chilled water filtered to 1 µm and either UV-disinfected or not for algal culture.

5 – Heated water filtered to 1 µm and either UV-disinfected or not for larval culture.

After filtration, all or part of the seawater may be pumped to a storage tank that may be made of either concrete or fibreglass. Use of a storage tank may be a matter of preference and many hatcheries do not have them. They are useful when water can only be obtained at a particular time, e.g. at high tide. Sometimes this method is used in areas where electrical power is unreliable to ensure a supply of seawater is always available. Sufficient water is pumped into the storage tank so it can supply the hatchery until the tank can be refilled. The tank is located at height so that the effect of gravity maintains a sufficient water flow through the hatchery. In other hatcheries, the salt water system is a flow-through system and water is pumped continuously through the hatchery for use where it is needed and then is discharged to waste. Recently, many hatcheries have installed recirculating or partial recirculating systems to reduce operating costs. This is particularly true if seawater is in short supply or if it has been heated or chilled. Recirculated water may be passed over biologically activated filters to remove metabolic wastes of the animals and held before it is reused. If the water has been heated or chilled it may be passed through heat exchangers to partially heat or chill incoming water and thus reduce energy costs.

All piping must be non-toxic, usually PVC (polyvinylchloride) schedule 40 or 80, although ABS or polyethylene pipes and fittings are also sometimes used as alternatives. The diameter of the pipes depends on water demand. In most hatcheries the main distribution lines within the hatchery are 50 mm diameter or less although the main intake pipes may be up to 15 cm diameter. The piping should be well supported and high enough off the ground so that it is out of the way but readily accessible for cleaning. Valves and outlets should be conveniently located. If the water is sufficiently filtered there should be little need to clean the lines frequently. Cleaning may be required periodically, hence, it is important to have clean-out ports or screw unions located conveniently so that the lines can be easily cleaned *in situ* or quickly dismantled for more thorough cleaning.

In most hatcheries in temperate areas there needs to be the capability to heat and sometimes to chill part of the seawater supply. There are commercial units available for this purpose and discussions and calculations on required capacity with dealers will ensure that an adequate supply at the required temperatures is available. Again, it is essential to ensure that surfaces of such units coming in contact with the seawater are non-toxic to bivalve larvae. Most commercially available heat exchange units utilize titanium as the heat transfer surface and this material is preferred by most hatcheries.

Hatchery managers may wish to sterilize (or more correctly, disinfect) all or part of the seawater before use, particularly if disease problems arise. Seawater can be sterilized with either UV (ultra-violet) light or ozone. Commercial units are available and simple calculations will determine the size of unit that is required. Commercial units are normally rated for their performance in sterilizing freshwater. In seawater situations where organic loadings and turbidity caused by colloidal materials are frequently higher than for freshwater, it is recommended that such units are used at half (or less) of the recommended flow rate for satisfactory performance. If UV-light sterilization is used, the water must be filtered to about 1 µm prior to sterilization since UV-light is readily absorbed by particles in the water reducing the efficiency of the unit. Filtration can easily be incorporated into a UV unit and many available units have both filters and the UV lamps combined.

Government regulations may exist in some areas that control the discharge of effluent from a hatchery. Before constructing a hatchery, government regulations controlling discharge of effluents should be reviewed and if they exist they must be followed.

Large floor drains sunk into the floors of wet areas are essential and should be located conveniently throughout the hatchery. Periodically large volumes of water must be discharged, e.g. when emptying tanks, and the drains must be able to handle such discharges.

Some hatcheries may wish to breed exotic species or strains or races of a species that do not occur locally. Depending on government regulations, this may entail installation of a quarantine facility to ensure that pests, parasites and diseases are not introduced with the exotic species or larvae accidentally escape into the natural environment. This will require a separate drainage system in the area of the hatchery designated for quarantine that empties into special holding tanks where the effluent can be sterilized with a strong hypochlorite solution. The sterilized water is then treated with thiosulphate to neutralize any residual chlorine before it is discharged back into the environment. Quarantine facilities may require a separate room to hold, condition and spawn adults. Drains from this room will also empty into the quarantine treatment tanks.

1.2.3 The physical plant

Careful thought should be given to hatchery design to permit convenient and efficient operations. The hatchery should be adaptable so that changes can be made readily without involving major rebuilding. In some hatcheries, tanks have been constructed of concrete and changes cannot be made easily. It is much better to have plastic or fibreglass tanks so they can be easily moved or changed if needed. Floors should be of concrete and have sufficient drains. All surfaces should be covered with a durable, mildew resistant finish to facilitate cleaning. Floor standing cabinets and storage units made from wood should be mounted on concrete plinths to prevent them being damaged by immersion in seawater. Where this is not possible, wood surfaces need to be painted with a good quality epoxy resin.

A hatchery has several areas that are all inter-related. For convenience they have been divided into algal culture, broodstock conditioning and spawning, larval rearing, juvenile culture and service areas (Figure 5).

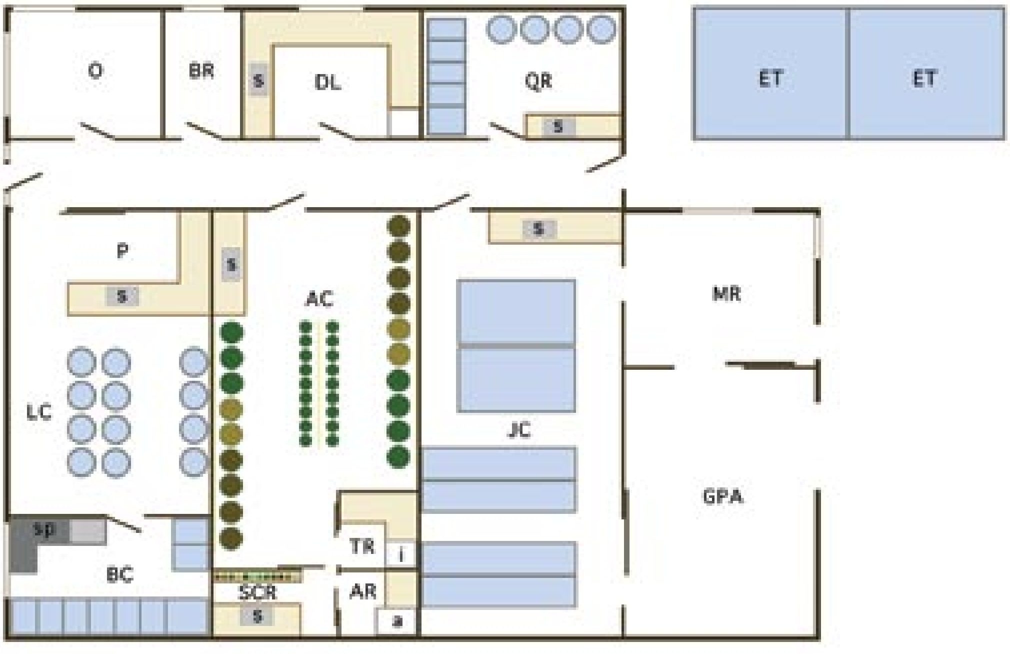

Figure 5: A generalized floor plan for a purpose-built bivalve hatchery (see the following text for explanation).

1.2.3.1 Algal culture facility

The success of a bivalve hatchery depends on the production of algae. Large quantities of high quality algae must be available when needed. It is a most important part of any hatchery and considerable thought should be given to providing a sufficient and efficient working area for this purpose (AC – Figure 5). Since algae are used in all phases of production, the facility should be located centrally and conveniently. Space required for algal culture depends partly on levels of production, methods of culture and whether algae will be raised entirely inside the hatchery with artificial illumination, or if it will be raised outside under natural light, or a combination of the two. A well ventilated greenhouse is required if algae is grown in natural light and this structure needs to be placed so as to obtain the maximum amount of sunlight. Shading may be need to protect younger, less dense cultures from strong sunlight.

A small room is required to maintain stock (also known as master) cultures of algae (TR). Dimensions vary but it can be as small as 2 x 3 m. The room should be insulated and the temperature kept cool. Shelving is needed with flourescent lights at the back to provide the light source. An air supply is also required. Test tubes with algal slants and small flasks with stock culture that are monospecific and axenic are kept in this room often in a refrigerated, illuminated incubator. Methods are described in a Part 3.

The next phase of culture uses stock cultures from the cool room and grows them in 4 l flasks and 20 l carboys against a bank of fluorescent lamps (SCR). This can be part of the main algal culture area or a small room off it. The space required depends on the number of species and amount of algae being produced. This area requires an air and carbon dioxide supply and needs to be kept at 15 to 18°C. Another adjacent small room (AR) houses an autoclave (a), which is used to heat sterilize medium for the small cultures. Some hatcheries use alternative methods to prepare culture medium and these are described in Part 3.

The size of the main algal culture area depends on the number of species being cultured and the amount of algae required. This area can occupy a substantial part of the hatchery. If most of the algae is raised inside the hatchery by the batch culture method then there must be sufficient space for a series of tanks that can measure up to 3-4 m in diameter and 2 m in depth. If the bag or tall cylinder culture methods are used the amount of floor area required can be reduced. Ballasts for fluorescent lamps used to illuminate cultures need to be of the "cool running" type or isolated in a separate area from which the heat they generate can be dissipated. This area should ideally be maintained at 15 to 20°C.

In many hatcheries, considerable portions of the algae, if not all, are raised in greenhouses. These can be stand-alone structures or attached to one side of the hatchery - preferably the south side in the Northern Hemisphere and northern side in the Southern Hemisphere, so as to receive as much sunlight as possible. The size of the greenhouse depends on the method of culture and quantities of algae that need to be produced.

Sufficient electrical power must be available for artificial lighting when natural sunlight is inadequate. Compressed air and carbon dioxide supplies are essential. There should be adequate ventilation or installed air-conditioning to maintain temperatures at or below 20°C on days when bright sunlight heats the facility. A generator will be required in areas where the electricity supply is unreliable and may be off for several hours or more at a time. Although survival of the algal cultures is not at risk in the absence of light for an hour or two, cultures need to be aerated. Diatoms will settle to the bottom of cultures without aeration and cultures may collapse.

1.2.3.2 Broodstock holding and spawning area

Space is required to hold and condition broodstock (BC – Figure 5). The amount of space needed depends in part on the number of species being held and whether some or most of the conditioning will be undertaken in the open environment rather than in the hatchery. Heated or chilled seawater may be required for this aspect of operation at certain times of the year. The ability to isolate tanks so that photoperiod can be adjusted is desirable since it has been shown that varying periods of light and dark can affect gonadal maturation.

Space is required for spawning trays (sp) but this can be part of the larval rearing area since the space is not require continuously. Spawning trays or dishes can be stored when not in use. Methods for broodstock conditioning, spawning and fertilization are described in Part 4.

1.2.3.3 Larval culture area

Another major part of the hatchery is occupied by the larval rearing facility (LC) and dimensions of this area depend on the scale of production. The space is occupied with tanks, the number needed depending on production levels and the techniques used to rear larvae. On the Pacific coast of North America the tendency has been to raise larvae at low densities of 2-3 per ml in large tanks that measure 3-4 m in diameter, 4-5 m in height and hold 40 000 to 50 000 l. In other hatcheries larvae are raised in smaller tanks of up to 5 000 l in volume at higher larval densities. A manager must decide on required production to meet market demand and the methodology that will be used to rear larvae when planning this part of the hatchery.

Larval rearing tanks are generally made of fibreglass or of a suitable plastic and should be thoroughly leached prior to use. Regardless of the size of tanks used, there should be large sunken floor drains to handle large volumes of water when the tanks are drained. A preparation area in the larvae culture room (P) is required for washing, grading, counting and measuring larvae and for accommodating the equipment used for these purposes. This area requires cupboards and shelves for the storage of equipment when not in use.

1.2.3.4 Juvenile culture area

Once mature larvae have set (settled and begun metamorphosis) they are moved to tanks in the juvenile culture room (JC) for culture until they are of sufficient size to transfer to nursery systems, which may be part of the hatchery or at another location. This is generally when the juveniles (known as spat) exceed 2 mm shell length. The size and types of tanks in terms of volume and surface area used for this purpose vary according to species.

Mature larvae are set in the hatchery or in outside (sometimes remote) facilities. When this procedure occurs within the hatchery it is generally done in the larval culture area, frequently directly in the larval tanks. Space for additional tanks may be required specifically for this process. Spat (early juveniles) are subsequently transferred to tanks systems in a separate area specifically for juvenile culture (JC). The size and types of tanks in terms of volume and surface area used for this purpose vary according to species. They may be upwellers, downwellers or tray systems of varying configuration and the juveniles are grown in these until they exceed 2 mm shell length. To grow spat to a larger size within the hatchery on cultured food is uneconomic since food requirement increases exponentially with size. If the nursery system is located outside the hatchery, sufficient space must be allotted for this operation.

Methods for the culture larvae are described in Part 5 and for spat in Part 6.

1.2.3.5 Other space requirements

Hatcheries dealing with broodstock from outside the immediate region or with exotic species may, as already mentioned, be required to quarantine stock and rear the progeny in isolation. Such hatcheries will include a quarantine room (QR) for this purpose, the effluent from which is discharged into treatment tanks (ET).

Other rooms include a dry laboratory (DL), office (O) and bathroom (BR). The dry laboratory is where algal transfers can be made (if no specific space is allocated elsewhere), chemicals weighed and mixed, microscopes kept for examining cultures, records maintained and for the storage of scientific equipment.

Static machinery such as the main pumps, sand filters and pre-filters (to remove particles down to 10 µm), seawater heating/chilling units, furnaces, the air ventilation system, air blowers/compressors, a standby generator for emergency power supply, together with electrical panels and control equipment, are housed in a soundproof machinery room (MR). Duplication of essential equipment is preferred in the event of electrical or mechanical failure. Compressed air is required in all phases of culture and carbon dioxide is required for algal culture. In many hatcheries the seawater intake pumps and sand filters are located in a separate pump house close to the point of intake and the final filtration of seawater may take place at the point of use rather than at a central, fine filtration unit.

Since storage is always an issue in a hatchery, it is useful to have a large general-purpose area (GPA) that can be used for storing materials and equipment, packing seed and as a workshop. Most of the working areas should be fitted with benches and sinks (s).

It is preferred that the various parts of the hatchery can be isolated in the event of a disease outbreak.

1.3 ECONOMIC CONSIDERATIONS

A bivalve hatchery is a business and like any other business it must be run efficiently and it must be economically viable. Government subsidies or grants may help offset costs particularly during initial stages of operation, but eventually the hatchery must stand on its own and be profitable. The economics of building and operating a bivalve hatchery will vary from business to business, from area to area and country to country but eventually all must turn a profit.

Hatcheries are expensive operations. Considerable capital is required to build a hatchery and finance operations. The owner must have sufficient working capital to carry on operations until income is generated. Before deciding to build a hatchery, one needs to carefully examine all facets of building and operating a hatchery and determine at what level a hatchery will be economically viable. Many costs need to be considered including purchase of the site, construction of the hatchery, installation of the seawater system, equipment needed for all phases of production, maintenance, supplies and utility overheads, loan repayments and the need for a trained staff.

Profitability can vary greatly with other factors including geographic area, the scale of the operation and whether it is part of a fully integrated bivalve culture operation.

In temperate areas a major operating cost is heating (and chilling) seawater, but this cost is generally avoided in tropical areas. This may influence location of a hatchery in temperate areas to sites where warm seawater exists at least for part of the year to help reduce heating costs.

Some hatcheries are small family operated ventures that only produce sufficient seed for their own culture needs. Such hatcheries are generally operated for only a few months a year, production is limited, and costs are much lower than for other larger hatcheries.

Large hatcheries may be part of a fully integrated bivalve culture operation or they may be in business only to supply seed. Where a hatchery is part of an integrated culture operation, the hatchery may be operated to simply break even and show no profit or may even operate at a small loss. Profits for the company are made in other phases of the culture operation. Where the hatchery exists only to produce seed to sell to other growers, a profit must be made solely on the hatchery operation. This emphasizes the fact that before building a hatchery one must make an accurate assessment of the market for whatever seed will be produced and not only the quantity of seed that can be sold but also the price people are willing to pay for seed.

Another consideration in operating a bivalve hatchery is that a critical level of production must be maintained to permit profitability. A hatchery cannot exist by simply producing a few thousand juveniles each year. The cost to do so is too high. In fact the costs associated with producing a few thousand juveniles are almost the same as producing several million – economies of scale apply. A manager must determine the critical level of production that needs to be attained to make the operation profitable and this again points to the necessity of knowing the extent and value of the market for the product.

Accurate records of costs, production and sales must be kept to assess whether the hatchery is being profitably run.

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Part 2

Basic bivalve biology: taxonomy, anatomy and life history

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2.1 TAXONOMY AND ANATOMY

2.1.1 Introduction

Some knowledge of bivalve biology is necessary to understand operations of a bivalve hatchery and to assist in solving problems that arise. It is not the intention here to give a detailed description of bivalve biology but to provide a brief resume of information pertinent to operations of a hatchery. There are several excellent texts on molluscan biology readily available and there are extensive reviews of groups and individual species of oysters, scallops, mussels and clams. The reader is directed to these publications at the end of this section for additional information.

Bivalves belong to the phylum Mollusca, a group that includes such diverse animals as chitons (chain shells), gastropods, tusk shells, cephalopods (squid and octopus) as well as clams, oysters, mussels and scallops. The phylum has six classes of which one is Lamellibranchia or Bivalvia. These animals are compressed laterally and the soft body parts are completely or partially enclosed by the shell, which is composed of two hinged valves. The gills or ctenidia of animals in this class are well developed organs, specialized for feeding as well as for respiration.

2.1.2 External anatomy

The most prominent feature of bivalves is the two valves of the shell that may or may not be equal and may or may not completely enclose the inner soft parts. They have a variety of shapes and colours depending on species. The valves are composed mostly of calcium carbonate and have three layers; the inner or nacreous layer, the middle or prismatic layer that forms most of the shell, and the outer layer or periostacum, a brown leathery layer which is often missing through abrasion or weathering in older animals.

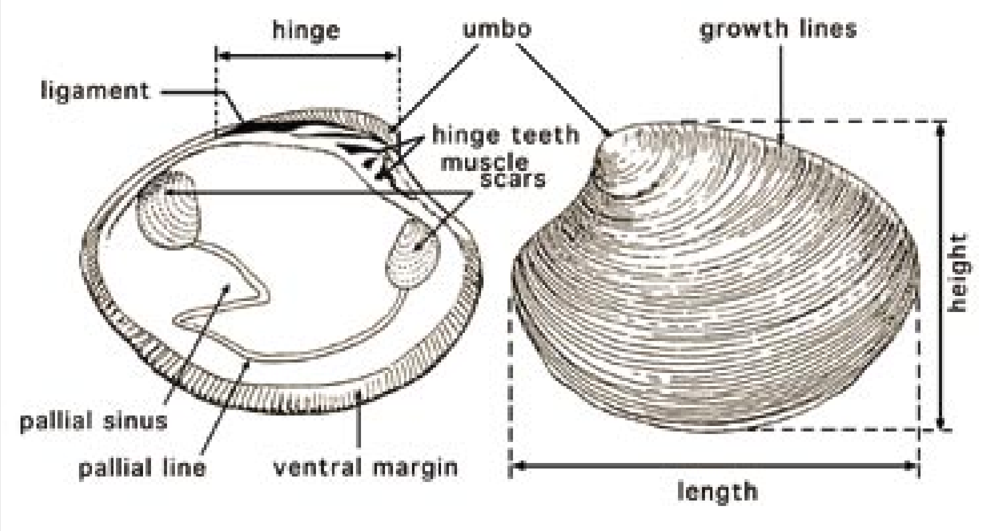

Figure 6: External and internal features of the shell valves of the hard shell clam, *Mercenaria mercenaria*. Modified from Cesari and Pellizzato, 1990.

Bivalves do not have obvious head or tail regions, but anatomical terms used to describe these areas in other animals are applied to them. The umbo or hinge area, where the valves are joined together, is the dorsal part of the animal (Figure 6). The region opposite is the ventral margin. In species with obvious siphons (clams), the foot is in the anterior-ventral position and the siphons are in the posterior area (Figure 7). In oysters the anterior area is at the hinge and in scallops it is where the mouth and rudimentary foot are located.

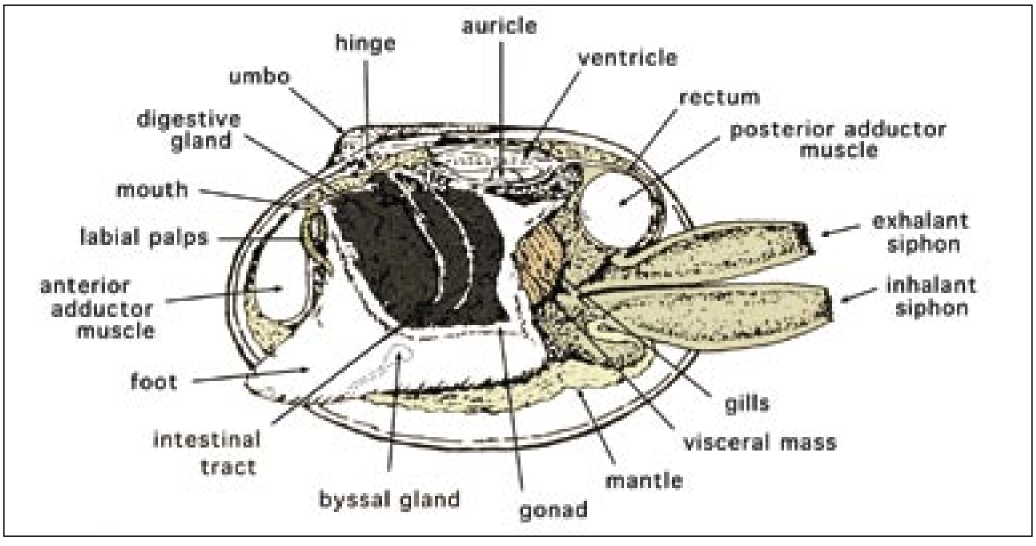

Figure 7: The internal, soft tissue anatomy of a clam of the genus *Tapes*. In this view, the uppermost gill lamellae have been removed to reveal the foot and other adjacent tissues. Modified from Cesari and Pellizzato, 1990.

2.1.3 Internal anatomy

Careful removal of one of the shell valves reveals the soft parts of the animals. The differences in general appearance of an oyster and scallop can be seen in Figure 8.

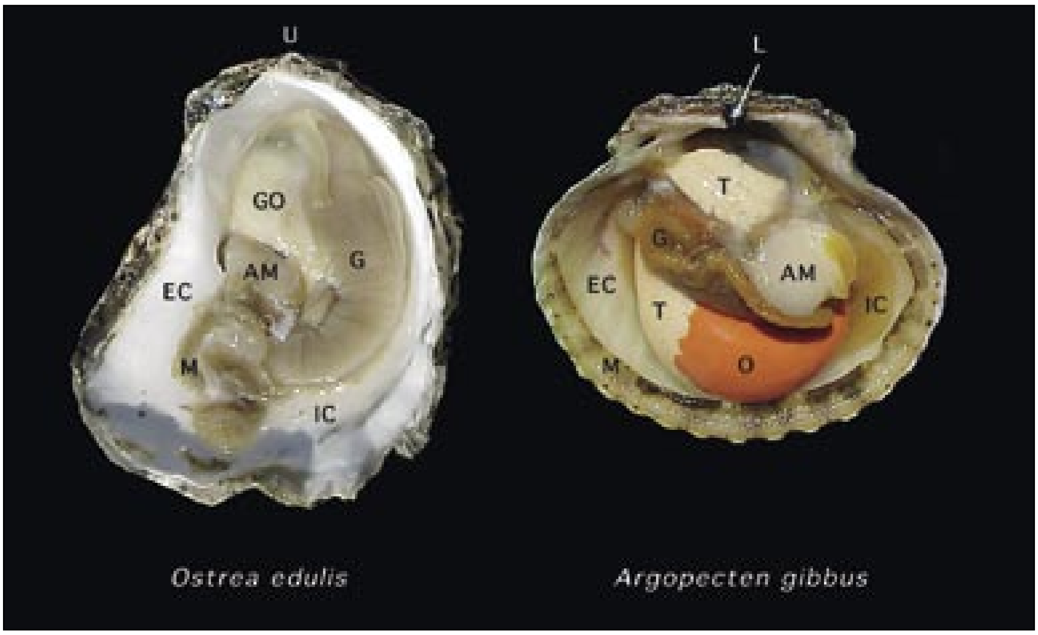

Figure 8: The soft tissue anatomy of the European flat oyster, *Ostrea edulis*, and the calico scallop, *Argopecten gibbus*, visible following removal of one of the shell valves. **Key**: **AM** – adductor muscle; **G** – gills; **GO** – gonad (differentiated as **O** – ovary and **T** – testis in the calico scallop); **L** – ligament; **M** – mantle and **U** – umbo. The inhalant and exhalant chambers of the mantle cavity are identified as **IC** and **EC** respectively.

Mantle

The soft parts are covered by the mantle, which is composed of two thin sheaths of tissue, thickened at the edges. The two halves of the mantle are attached to the shell from the hinge ventral to the pallial line but are free at their edges. The thickened edges may or may not be pigmented and have three folds. The mantle edge often has tentacles; in clams the tentacles are at the tips of the siphon. In species such as scallops the mantle edge not only has tentacles but also numerous light sensitive organs – eyes (Figure 9).

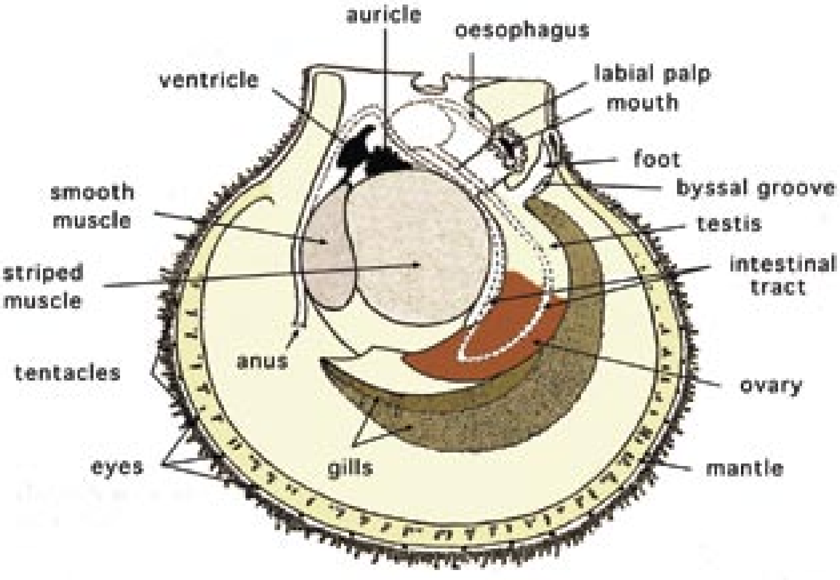

Figure 9: The internal, soft tissue anatomy of a hermaphroditic scallop.

The main function of the mantle is to secrete the shell but it also has other purposes. It has a sensory function and can initiate closure of the valves in response to unfavourable environmental conditions. It can control inflow of water into the body chamber and, in addition, it has a respiratory function. In species such as scallops, it controls water flow into and out of the body chamber and hence movement of the animal when it swims.

Adductor muscle(s)

Removal of the mantle shows the underlying soft body parts, a prominent feature of which are the adductor muscles in dimyarian species (clams and mussels) or the single muscle in monomyarian species (oysters and scallops). In clams and mussels the two adductor muscles are located near the anterior and posterior margins of the shell valves. The large, single muscle is centrally located in oysters and scallops. The muscle(s) close the valves and act in opposition to the ligament and resilium, which spring the valves open when the muscles relax. In monomyarian species the divisions of the adductor muscle are clearly seen. The large, anterior (striped) portion of the muscle is termed the "quick muscle" and contracts to close the valves shut; the smaller, smooth part, known as the "catch muscle," holds the valves in position when they have been closed or partially closed. Some species that live buried in the substrate (e.g. clams) require external pressure to help keep the valves closed since the muscles weaken and the valves open if clams are kept out of a substrate in a tank.

Gills

The prominent gills or ctenidia are a major characteristic of lamellibranchs. They are large leaf-like organs that are used partly for respiration and partly for filtering food from the water. Two pairs of gills are located on each side of the body. At the anterior end, two pairs of flaps, termed labial palps, surround the mouth and direct food into the mouth.

Foot

At the base of the visceral mass is the foot. In species such as clams it is a well developed organ that is used to burrow into the substrate and anchor the animal in position. In scallops and mussels it is much reduced and may have little function in adults but in the larval and juvenile stages it is important and is used for locomotion. In oysters it is vestigial. Mid-way along the foot is the opening from the byssal gland through which the animal secretes a thread-like, elastic substance called "byssus" by which it can attach itself to a substrate. This is important in species such as mussels and some scallops enabling the animal to anchor itself in position.

Digestive system

The large gills filter food from the water and direct it to the labial palps, which surround the mouth. Food is sorted and passed into the mouth. Bivalves have the ability to select food filtered from the water. Boluses of food, bound with mucous, that are passed to the mouth are sometimes rejected by the palps and discarded from the animal as what is termed "pseudofaeces". A short oesophagus leads from the mouth to the stomach, which is a hollow, chambered sac with several openings. The stomach is completely surrounded by the digestive diverticulum (gland), a dark mass of tissue that is frequently called the "liver". An opening from the stomach leads to the much-curled intestine that extends into the foot in clams and into the gonad in scallops, ending in the rectum and eventually the anus. Another opening from the stomach leads to a closed, sac-like tube containing the crystalline style. The style is a clear, gelatinous rod that can be up to 8 cm in length in some species. It is round at one end and pointed at the other. The round end impinges on the gastric shield in the stomach. It is believed it assists in mixing food in the stomach and releases enzymes that assist in digestion. The style is composed of layers of mucoproteins, which release digestive enzymes to convert starch into digestible sugars. If bivalves are held out of water for a few hours the crystalline style becomes much reduced and may disappear but it is reconstituted quickly when the animal is replaced in water.

Circulatory system

Bivalves have a simple circulatory system, which is rather difficult to trace. The heart lies in a transparent sac, the pericardium, close to the adductor muscle in monomyarian species. It consists of two irregular shaped auricles and a ventricle. Anterior and posterior aorta lead from the ventricle and carry blood to all parts of the body. The venous system is a vague series of thin-walled sinuses through which blood returns to the heart.

Nervous system

The nervous system is difficult to observe without special preparation. Essentially it consists of three pairs of ganglia with connectives (cerebral, pedal and visceral ganglia).

Urogenital system

Sexes of bivalves can be separate (dioecious) or hermaphroditic (monoecious). The gonad may be a conspicuous, well defined organ as in scallops or occupy a major portion of the visceral mass as in clams. The gonad is generally only evident during the breeding season in oysters when it may form up to 50% of the body volume. In some species such as scallops, the sexes can be readily distinguished by eye when the gonad is full since the male gonad is white in colour and the female is red, even in hermaphroditic species. Colour of the full gonad may distinguish the sexes in some species such as mussels. In other species, microscopic examination of the gonad is required to determine the sex of the animal. A small degree of hermaphrodism may occur in dioecious species.

Protandry and sex reversal may occur in bivalves. In some species there is a preponderance of males in smaller animals indicating that either males develop sexually before females or that some animals develop as males first and then change to females as they become larger. In some species, e.g. the European flat oyster, *Ostrea edulis*, the animal may spawn originally as a male in a season, refill the gonad with eggs and spawn a second time during the season as a female.

The renal system is difficult to observe in some bivalves but is evident in such species as scallops where the two kidneys are two small, brown, sac-like bodies that lie flattened against the anterior part of the adductor muscle. The kidneys empty through large slits into the mantle chamber. In scallops, eggs and sperm from the gonads are extruded through ducts into the lumen of the kidney and then into the mantle chamber.

2.2 LIFE HISTORY

2.2.1 Gonadal development and spawning

In most bivalves sexual maturity is dependent on size rather than age and size at sexual maturity depends on species and geographic distribution. Production of eggs and sperm is termed gametogenesis and size of the bivalve along with temperature and quantity and quality of food are undoubtedly important in initiating this process. The gonad is composed of many-branched, ciliated ducts from which numerous sacs, termed follicles, open. Gametes arise by proliferation of germinal cells that line the follicle wall. The gonad undergoes continuous development until it becomes fully mature but this development has been divided into several stages for convenience, e.g. resting, developing, mature, partially spawned and spawned. When the gonads or gonadal tissue are fully mature they are very evident and form a significant portion of the soft parts of the animal. Gonaducts that will carry the gametes to the body chamber develop, enlarge and are readily observed in the gonad. At this time the animal is frequently referred to as being ripe.

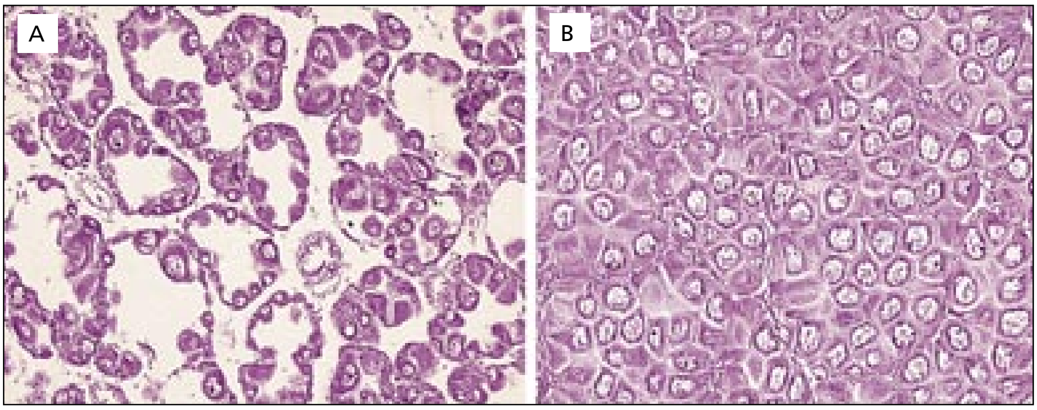

Figure 10: Photomicrographs of histological sections through the ovary of the scallop, *Argopecten gibbus*, during gametogenesis. To the left (**A**), developing eggs can be seen lining the walls of the numerous follicles. The right hand photograph (**B**) shows the follicles filled with mature eggs (courtesy: Cyr Couturier and Samia Sarkis).

Several methods have been used to determine if a bivalve is ripe and ready to spawn. The most accurate method is to make histological slides of the gonad (Figure 10) but this is costly, time consuming and the animal must be sacrificed. Making smears of the gonad or extracting small samples of the gonad from a few individuals of a stock and examining them microscopically is an alternative and the most frequently used technique. In scallops, the gonadal index (weight of the gonad divided by the weight of the soft parts, multiplied by 100) is sometimes used. A rigid routine is generally followed in hatcheries to condition adults for spawning and with practice, most hatchery managers quickly develop the ability to know if the animal is ripe and ready to spawn by examining the gonad macroscopically.

Bivalves that reach the size of sexual maturity and spawn for the first time are sometimes referred to as virgins. Although these animals attain sexual maturity, the number of gametes produced is limited and sometimes not all are viable. During subsequent spawning the number of gametes produced greatly increases.

The period of spawning in natural populations differs with species and geographic location. Spawning may be triggered by several environmental factors including temperature, chemical and physical stimuli, water currents or a combination of these and other factors. The presence of sperm in the water will frequently trigger spawning in other animals of the same species. Some bivalve species in tropical environments have mature gametes throughout the year and limited spawning may occur continuously during the year. In temperate areas, spawning is usually confined to a particular time of the year. Many bivalves undergo mass spawning and the period of spawning may be brief. Virtually the entire contents of the gonad are released over a short period during spawning activity. Other species of bivalves have a protracted spawning period and it may extend over a period of weeks. These species are sometimes referred to as dribble spawners. Limited spawning occurs over a protracted period with one or two major pulses during this time. In some species there may be more than one distinct spawning in a year. In hermaphroditic species, spawning is timed so that either the male or female part of the gonad spawns first. This minimizes the possibility of self-fertilization.

In most bivalve species of commercial interest, gametes are discharged into the open environment where fertilization occurs. Sperm is discharged in a thin, steady stream through the exhalent opening or exhalent siphon. Discharge of eggs is more intermittent and they are emitted in clouds from the exhalent opening or siphon. In species such as scallops or oysters, females frequently clap the valves to expel the eggs. This may be done to clear eggs lodged on the gills. After spawning the gonads of many species are emptied and it is impossible to macroscopically distinguish sex in individuals. The animal is then said to be in the resting stage. In dribble spawners, the gonad may never be completely emptied.

Some bivalves, e.g. the European flat oyster, are larviporous and the early stages of larval development occur in the inhalant chamber of the mantle cavity of the female-phase oyster. Eggs when spawned are passed through the gills and are retained in the mantle chamber. Sperm is taken in through the inhalant opening. The length of time larvae are held in the mantle chamber and subsequently the length of time larvae are free living in surface waters varies with species. In some genera, e.g. *Tiostrea*, larvae may only be part of the plankton for as little as one day.

Occasionally, particularly in temperate areas, spawning may not occur in some years. This can be a consequence of several factors but probably mostly is related to water temperatures, which remain too low to trigger spawning. When this occurs in oysters, the eggs and sperm may be reabsorbed into the gonadal tissue, broken down and stored as glycogen. In clams and scallops the gonad may remain in a ripe condition until the next year.

2.2.2 Embryonic and larval development

These topics are covered more fully in later sections, but a brief description is given here for continuity. Larval development is similar whether initial development occurs in the mantle chamber of the female or completely in the open environment.

Eggs undergo meiotic division at fertilization to reduce the number of chromosomes to a haploid number before the male and female pronuclei can fuse to form the zygote. Two polar bodies are released during meiotic division and when apparent, indicate successful ferilization. Cell division begins and within thirty minutes after fertilization the egg divides into the two-celled stage. The eggs are heavier than water and sink to the bottom of the tank where cell division continues.

The time taken for embryonic and larval development is species specific and temperature dependent (Figure 11). Within 24 hours the fertilized egg has passed through the multi-celled blastula and gastrula stages and in 24 to 36 hours has developed into a motile trochophore. Trochophores are somewhat oval in shape, about 60-80 µm in size and have a row of cilia around the middle with a long apical flagellum and these permit them to swim.

The early larval stage is referred to as the straight-hinge, "D" or Prodissoconch I stage. Shell length of the initial straight-hinge stage varies with species but it is generally 80-100 µm (larger in larviparous oysters). The larva has two valves, a complete digestive system and an organ called the velum that is peculiar to bivalve larvae. The velum is circular in shape and can be protruded from between the valves. It is ciliated along its outer margin and this organ enables the larva to swim but only well enough to maintain itself in the water column. When the larva swims through the water column the velum collects phytoplankton upon which the larva feeds.

Larvae continue to swim, feed and grow and within a week the umbones, which are protuberances of the shell near the hinge, develop. As larvae continue to grow, the umbones become more prominent and the larvae are now in the umbone or Prodissoconch II stage. Prodissoconch II stage larvae have distinct shapes and with practice it is possible identify larvae of different bivalve species in the plankton. This has been used by biologists to forecast oyster sets in the natural environment for the industry. Duration of the larval stage varies with species and environmental factors such as temperature but it can be 18-30 days. Size at larval maturity also varies with species and can be 200-330 μm.

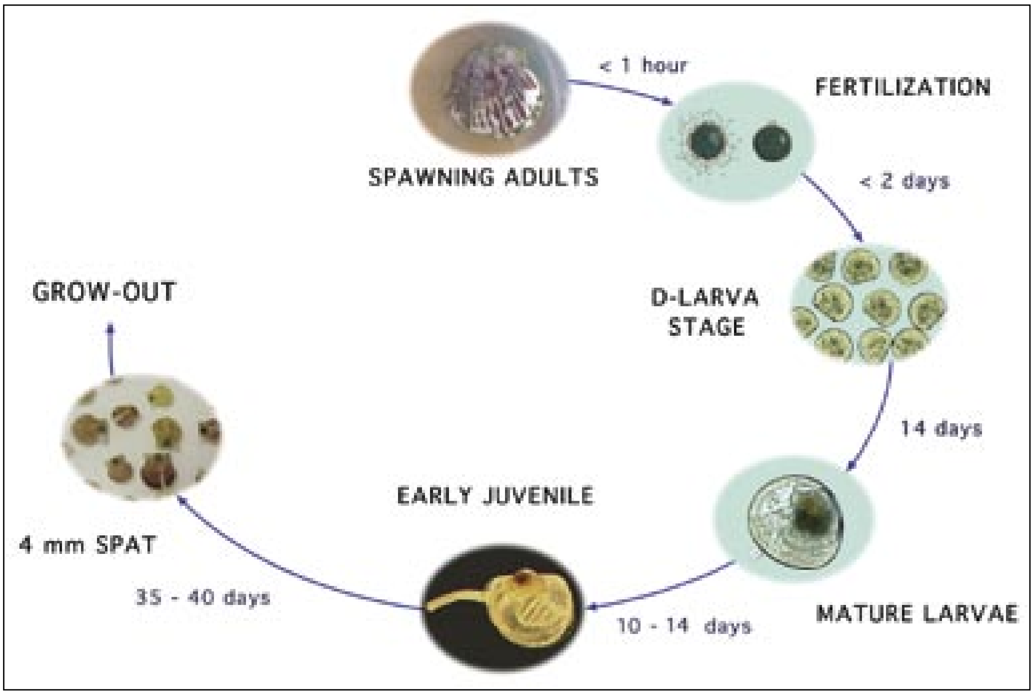

Figure 11: Representation of the developmental stages of the calico scallop, *Argopecten gibbus*, which take place within a hatchery. The duration of the period between the various stages is given in hours or days for this particular species and may differ for other species of bivalves.

When larvae approach maturity, a foot develops and gill rudiments become evident. Small dark circular dots, the eye-spots, develop near the centre of each valve of some species. Between periods of swimming activity, larvae settle and use the foot to crawl on a substrate. When a suitable substrate is found the larva is ready to metamorphose and begin its benthic existence. Mature oyster larvae secrete a small drop of cement from a gland in the foot, roll over and place the left valve in it. They remain attached in that location for the rest of their lives. In other species, the larva secretes byssus from the byssal gland in the foot and this serves as a temporary holdfast to attach to a substrate. The larva is now ready to metamorphose.

2.2.3 Metamorphosis

Metamorphosis is a critical time in the development of bivalves, during which the animal changes from a swimming, planktonic to a sedentary benthic existence. Considerable mortalities can occur at this time both in nature and in hatcheries. The subject is dealt with in detail later since it is an important aspect in the hatchery production of juvenile bivalves.

2.2.4 Feeding

Bivalves are filter feeders and feed primarily on phytoplankton – microscopic plant life. In juveniles and adults, the ctenidia, or gills, are well developed and serve the dual purpose of feeding and respiration. The ctenidia are covered with cilia – tiny vibrating hairs – whose concerted and co-ordinated beat induces a current of water. When resting on or in a substrate, water is drawn into the animal through the inhalant opening or the inhalant siphon, through the gills and then is returned to the surrounding water through the exhalant opening or siphon. The gills collect plankton and bind it to mucous. Strands of food-laden mucous are passed anteriorly by means of ciliary action along special grooves on the gills to the labial palps whose role is to assist in directing food into the mouth. Bivalves can exercise some selection of their food and periodically the palps reject small masses of food, pseudofaeces, that are expelled from the mantle cavity, often by the vigorous "clapping" together of the shell valves.

What constitutes optimum foods for bivalves remains largely unknown however phytoplankton undoubtedly forms the major portion of the diet. Other sources of food may be important such as fine particles of non-living organic material (detritus) along with associated bacteria and also dissolved organic material.

2.2.5 Growth

Only general statements can be made about growth in juveniles and adults since it varies greatly between species, geographic distribution, i.e. climate, location in the subtidal or intertidal zones, as well as differences in individuals and in their genetic make up. Growth can also vary greatly from year to year and in temperate areas there are seasonal patterns in growth.

Growth can be measured in bivalves by different methods including increments in shell length or height, increases in total or soft body weight, or a combination of all of these factors. In tropical areas growth can vary with season, being faster during or after rainy periods when nutrients are washed into the ocean leading to increased production of phytoplankton. In temperate areas, growth is generally rapid during spring and summer when food is abundant and water temperatures are warmer. It virtually ceases in winter, resulting in annual checks in the shell. These winter checks can be used to age some bivalves. Some species are short lived but others may live to over 150 years.

In culture operations the important considerations in bivalve growth are length of time taken to grow to sexual maturity and to market size. The goal of bivalve culture is to grow bivalves to commercial size as quickly as possible to make the operation as economically attractive as possible.

2.2.6 Mortalities

Bivalves in the larval, juvenile and adult stages can die from a variety of causes, which can be environmental or biological in origin. The subject is much too large to consider in detail here but a brief synopsis is given to highlight a number of pertinent points, which could be important in hatchery operations.

The physical environment can cause severe mortalities to bivalves in all three stages. Too high temperatures or prolonged periods of cold temperatures can be lethal to bivalves as can be sudden swings in temperature. Severe extremes in salinities, particularly low salinities after periods of heavy rain or run off from melting snow, can also cause extensive mortalities. Heavy siltation can smother and kill juveniles and adults.

Pollution, particularly industrial pollution, can cause extensive mortalities in juvenile and adult bivalves. Both industrial and domestic pollution can be problems for hatchery operations and must be avoided. Domestic pollution can increase organic and bacterial loads in water as well as contributing a wide range of potentially toxic materials. Little is known of the combined effects of sub-lethal levels of the wide range of organic and organo-metallic compounds of man-made origin that may be present in such effluents.

Bivalves in the larval, juvenile and adult stages are preyed upon by a wide variety of animals that can cause severe mortalities. In the natural environment plankton feeders probably consume large quantities of larvae. In hatcheries, predation is largely a non-issue since the water used is filtered and any predators are removed.

Bivalves are hosts to parasites that can cause mortalities, particularly in the adult stage. Shell boring worms, *Polydora* sp., and sponges burrow into the shells and weaken them, thus causing mortalities.

Probably the major cause of mortalities in bivalves, particularly of larvae and juveniles in hatcheries, is disease. Considerable research effort has been expended in studying bivalve diseases and trying to develop methods to control them.

Diseases can be devastating to adult bivalves as witness the demise of some populations in the world. A few examples include,

Dermocystidium:
a fungal disease of bivalves caused by *Perkinsus marinus;*

Delaware Bay Disease (MSX):
a disease caused by the haplosporidian protozoan, *Haplosporidium (Minchinia) nelsoni*;

SSO (seaside organism disease):
a disease caused by the haplosporidian protozoan, *Haplosporidium costale*, (which together with *H. nelsoni* has decimated large populations of Virginia oysters on the Atlantic coast of the USA and now extends northwards into Atlantic Canada).

Aber Disease:
A disease caused by the protozoan, *Marteilia refringens;*

Bonamiasis (Haemocytic Disease):
A disease caused by the microcell parasite, *Bonamia ostreae*;
(Aber disease and Bonamiasis have resulted in the virtual demise of the European oyster in some parts of Europe).

Although considerable work has been carried out on these diseases, no practical methods have been developed to control them and restore oyster populations to previous levels. The severity of these diseases points to the care that must be taken when transporting adult bivalve stock into a hatchery.

In hatcheries it appears that diseases which do occur are caused by bacteria and not by protozoans. Bacteria are present to some degree in both algal and larval cultures. Indeed, bacteria may form an important part of the diet of larvae. However, periodically, large groups of larvae will die suddenly and an entire culture is lost. High bacterial counts are almost always associated with such large-scale mortalities. Bacteria may cause mortalities (pathogenic) or they may be simply present as opportunistic bacteria (saphrophytic), feeding on the dying larvae. Bacteria that cause diseases largely belong to the genus *Vibrio* sp. and every precaution must be taken to prevent them from causing epidemics in hatcheries. The best method to prevent such epidemics is to observe strict hygienic procedures and ensure that the larvae are well fed with high quality food. Larvae should be inspected regularly. If a disease occurs or is suspected, tanks and equipment should be sterilized with a bleach solution and rinsed well with freshwater. To protect larvae from further contamination, tanks should be refilled with UV-irradiated or ozone treated seawater. Use of antibiotics to control diseases is largely avoided in hatcheries. They are expensive and add to cost of operations and also there is the fear of a strain of bacteria developing that will be resistant to the antibiotics, which could lead to even more severe disease problems in the future.

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